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FIELD AND LABORATORY TECHNIQUES FOR EVALUATING HEMLOCK SAWFLY INFESTATIONS

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by

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ABSTRACT

The hemlock sawfly, Neodiprion tsugae, *causes much defoliation of western hemlock in localized areas in southeastern Alaska. As the intensity of forest resource management increases, there will be increasing demands placed on forest pest management specialists to assess the course of infestations. This is already apparent in high value stands such as in recreation areas, near population centers, and in high-risk, overmature stands. This paper discusses techniques that will permit pest management personnel to determine trends in sawfly populations which could influence resource management decisions. Procedures for sampling and handling all stages of the sawfly and methods for determining population parameters such as larval instar composition, survival, sex ratio, fecundity, and classes of mortality are presented. Figures, descriptions, and a key are given to help the user diagnose and identify specific sources of mortality.*

Keywords: *Hemlock sawfly,* Neodiprion tsugae, *insect damage (-forest, population sampling (-forest protection.*

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INTRODUCTION

The hemlock sawfly, *Neodiprion tsugae* Midd. (fig. 1), is a major defoliator of western hemlock, *Tsuga heterophylla* (Raf.) Sarg. It feeds on previous year needles, and current year needles are avoided. Often, needles are only partially consumed, with needle stubs and midribs remaining. Larvae feed in colonies (fig. 2) and when mature spin tan, capsule-shaped cocoons (fig. 3) on host or understory vegetation.

Heavily defoliated stands (fig. 4) usually recover in a few years, but a few of the most severely defoliated trees die and some additional trees are top-killed (table 1). Radial growth

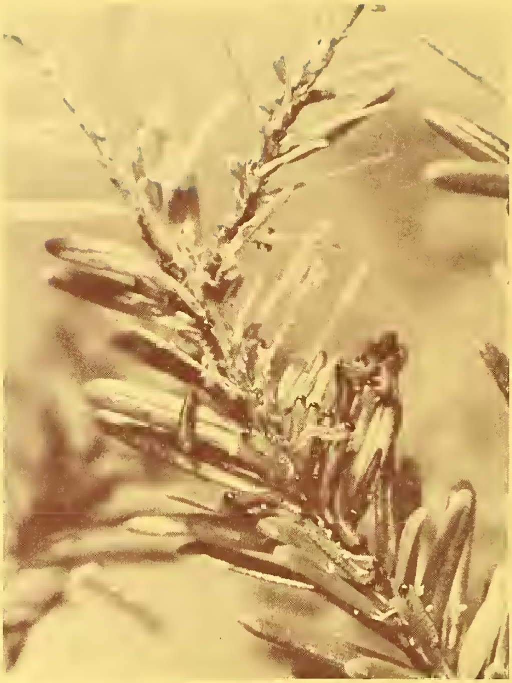

Figure 2.--Feeding colony of early instar hemlock sawfly larvae. Note character of needles which have been fed upon.

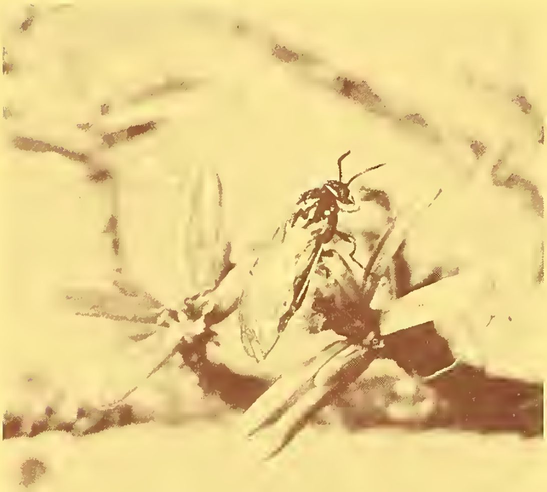

Figure 1.--Adult female hemlock sawfly on foliage of western hemlock.

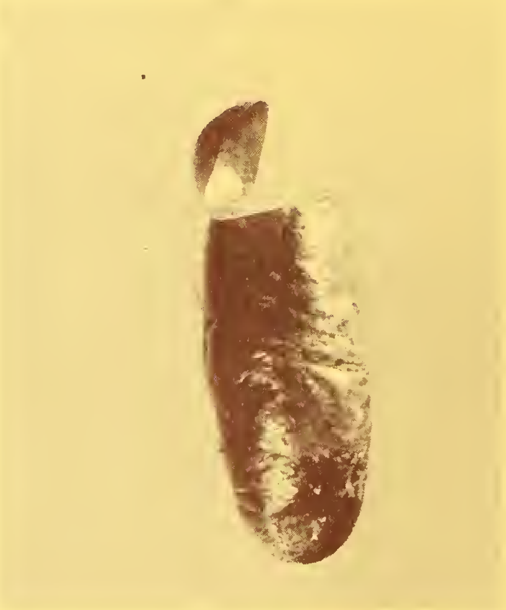

Figure 3.--Hemlock sawfly cocoon showing the evenly cut margin of cap resulting from normal sawfly emergence.

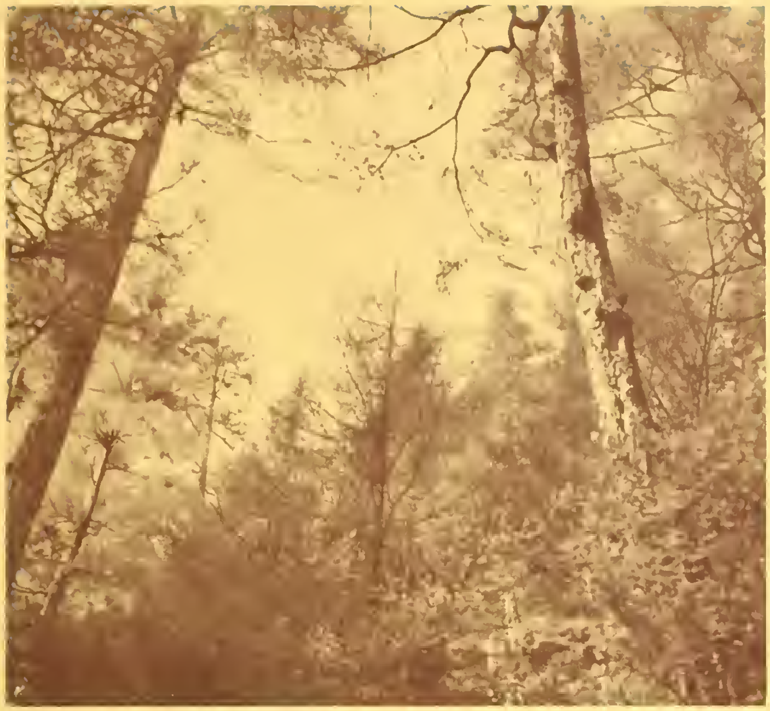

Figure 4.--Severe defoliation of mature western hemlock caused by hemlock sawfly.

Table 1.--*Western hemlock damage and recovery from 2 years of heavy sawfly defoliation on three 1/5-acre plots near Todd, Alaska*

Degree of defoliation, September 1967[1]	Mean d.b.h.	Total	Condition of trees, August 1970			
			Apparently healthy	Thin crown	Top kill	Dead
	Inches	- - - - - - - -	Number of trees	- - - - - - -		
None to light	16.2	16	15	0	1	0
Moderate	19.1	18	16	0	2	0
Heavy	21.8	26	15	3	8	0
Severe	16.5	7	0	3	2	2
Total	19.2	67(100%)	46(69%)	6(9%)	13(19%)	2(3%)

[1] Year that sawfly outbreak collapsed.

in most defoliated trees is temporarily reduced (fig. 5).

Most sawfly infestations in southeast Alaska occur south of Frederick Sound. Major population regulators are a larval fungus disease, most common during wetter-than-average summers, and three parasite species, most abundant during drier-than-average summers. Between 1952 and 1974 most declining sawfly population trends south of Frederick Sound occurred after two consecutive wetter-than-average summers (fig. 6). Unusually cold summer and fall weather delays larval development and inhibits oviposition.

This paper provides field and laboratory techniques required for pest management entomologists to determine whether a sawfly population is likely to increase or decrease. No recommendations are given for suppression because no pesticides have been field-tested against the sawfly, and materials and techniques will change over time.

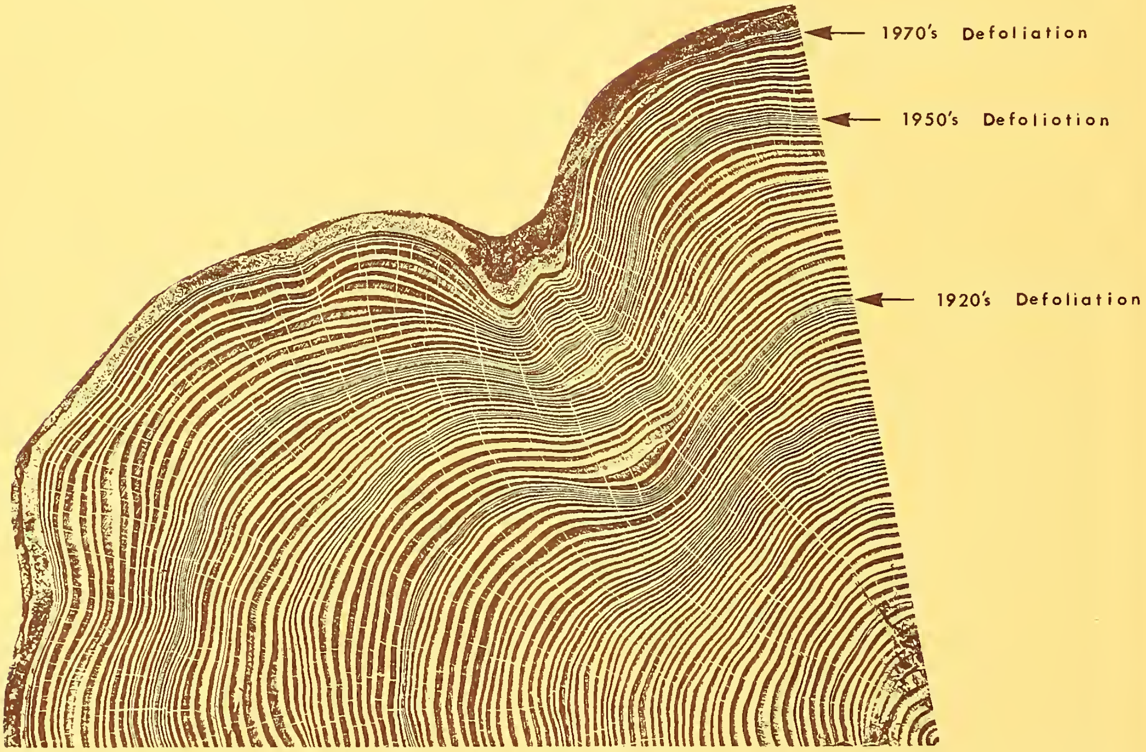

Figure 5.--Western hemlock stem section showing reduced ring widths as a result of defoliation.

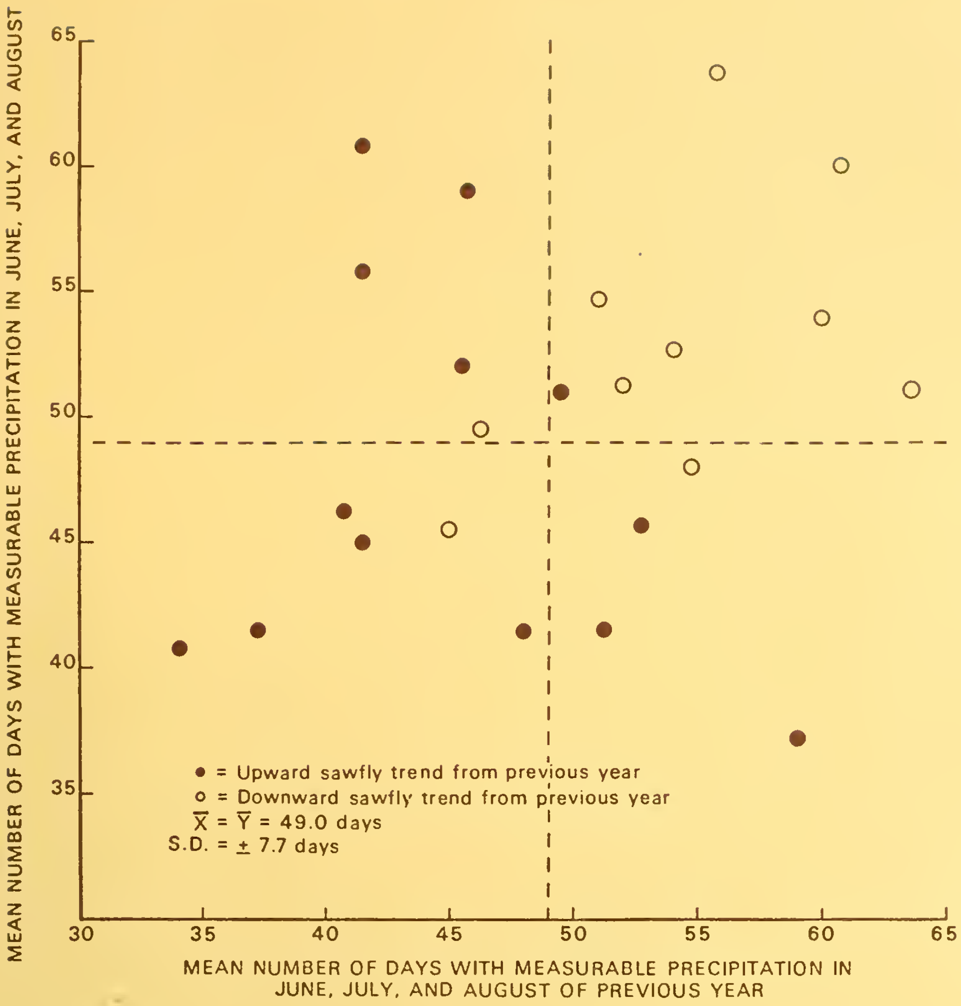

Figure 6.--Hemlock sawfly population trends south of Frederick Sound relative to frequency of precipitation during the larval periods of paired consecutive years, 1951-74 (Hard, in press).

LARVAL SAMPLING

A simple technique for collecting large numbers of larvae is to place a drop cloth beneath infested hemlock foliage and strike the branches sharply with a pole. Using a moistened camel's hair paint brush for young larvae and forceps for older larvae the dislodged larvae are then transferred from the drop cloth to vials containing 75-percent ETOH or KAAD.[1]

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[1] A mixture composed of 1 part kerosene, 10 parts 95-percent ethyl alcohol (ETOH), 2 parts glacial acetic acid, and 1 part dioxane.

Larval instar composition of a collection can be determined by measuring individual head capsule widths using a dissecting stereomicroscope fitted with an ocular micrometer. Larval head capsule widths are compared against a standard (fig. 7), and percentages of larvae in the various instars are computed. Most male larvae have four feeding instars, and most female larvae have five. Head capsule widths of prepupal instars are not significantly different from widths of the last feeding instar.

It is often important to know the sex ratios of a series of larval collections from an infestation in order to determine whether change has occurred as a result of sex-differentiated mortality. Sex of individual larvae can be determined in several ways, especially in the later instars. One technique, which can also be used to determine the sex of live larvae, is to examine the underside of the larval abdomen (fig. 8) under a dissecting microscope. Ovipositor bud plates and associated structures occur only on females.

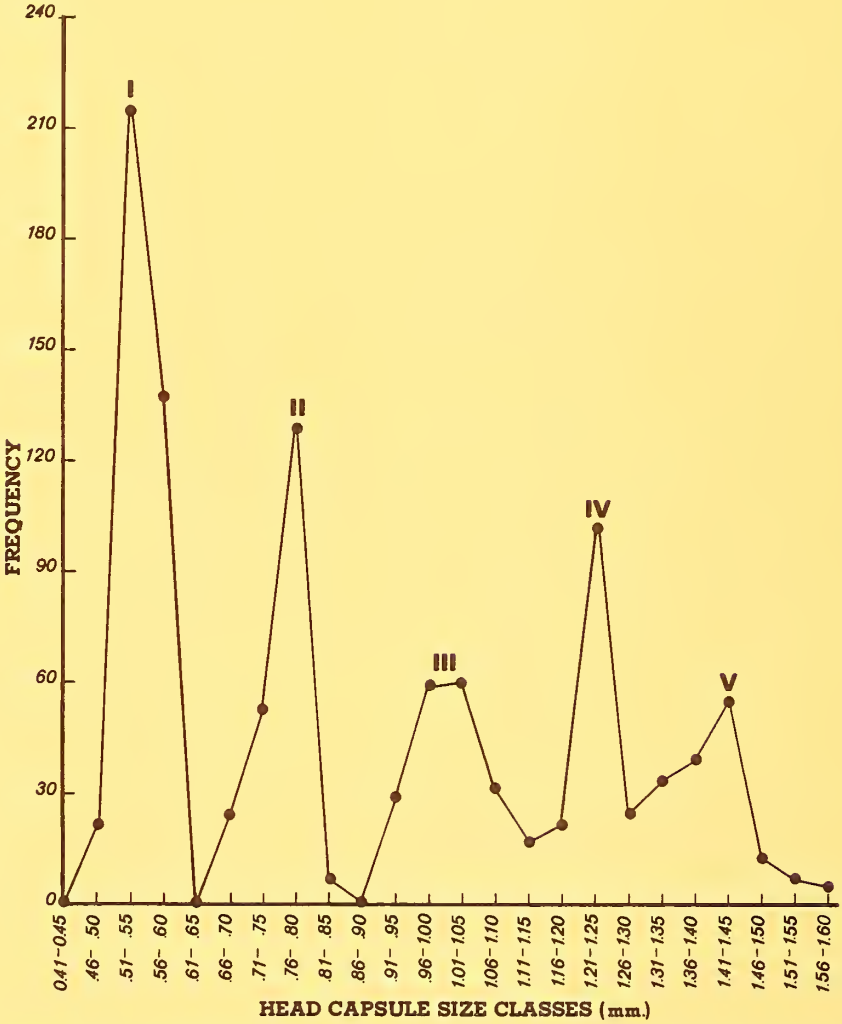


Figure 7.--Hemlock sawfly larval head capsule widths (Hard and Schmiege 1968).

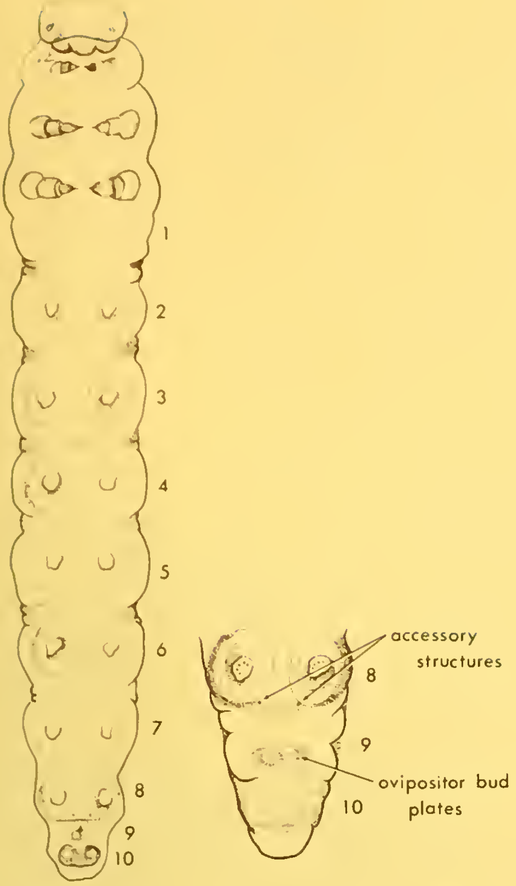


Figure 8.--Ventral view of hemlock sawfly larva and enlarged view of terminal segments showing position of ovipositor bud plates and accessory structures.

Another technique is removing and examining the gonads, which are located internally between the fifth and sixth abdominal segments. Gonads of both sexes are more opaquely white and less translucent than the surrounding fat body (fig. 9). Ovaries resemble bunches of bananas (fig. 10A); testes resemble cauliflowers (fig. 10B) and are usually larger than ovaries.

The major biotic mortality factor of sawfly larvae in Alaska is a fungus, *Entomophthora sphaerosperma* Fres. Recently killed larvae are brown and rigid but are soon covered with a

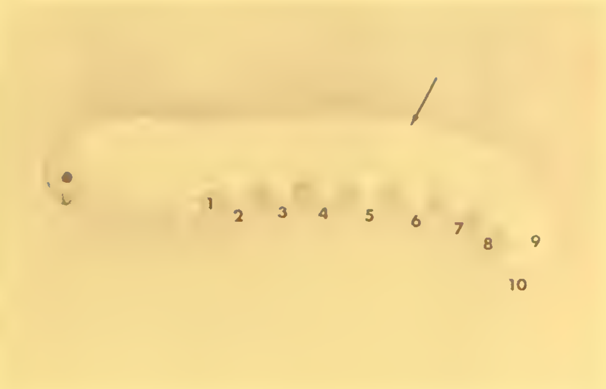


Figure 9.--Side view of prepupal sawfly larva showing position of gonad.

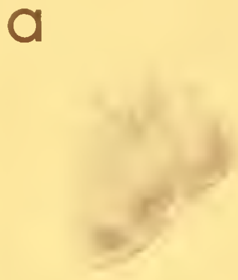
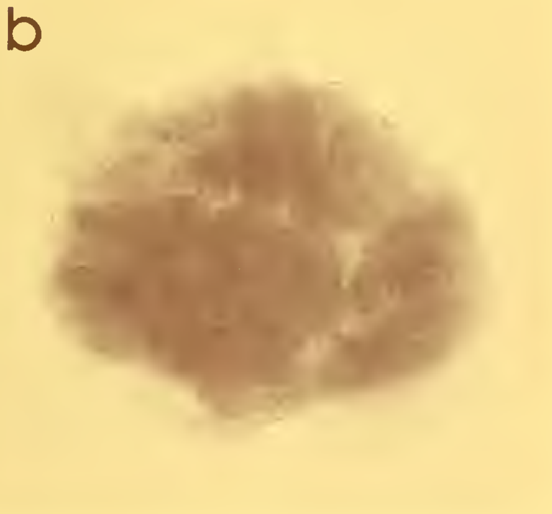


Figure 10.--Larval gonads: A, ovary (0.25 x 0.45 mm); and B, teste (0.45 x 0.59 mm).



light brown, velvety fungal mat (fig. 11). The disease usually becomes most apparent in late larval populations in early to middle August when most larvae are in the last feeding or prepupal instars. Head capsule widths of preserved larvae killed by the fungus can be measured in the laboratory to determine approximate sex ratio by comparing individual widths with figure 7. Those widths that fall in the fifth instar represent females.

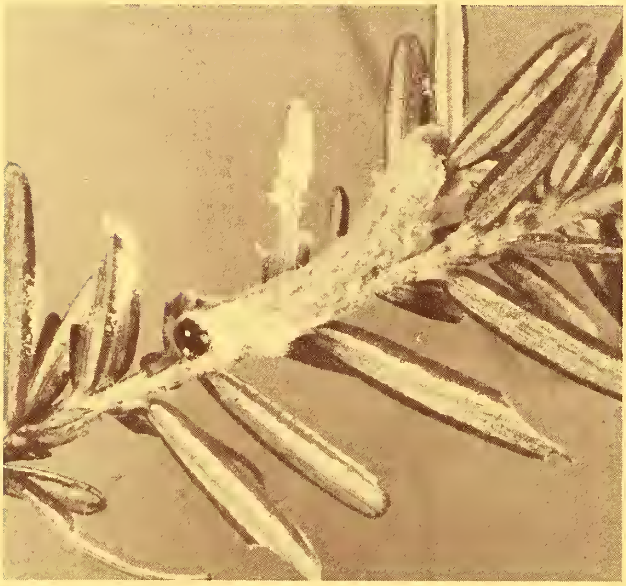


Figure 11.--Hemlock sawfly larva killed by Entomophthora sphaerosperma.

COCOON SAMPLING

During the years 1969 through 1973 in the southern Panhandle, cocoon formation began in the second or third week in August. Field studies showed that by mid-October at least 40 percent of the current year's cocoons had fallen from the foliage and twigs where they were formed. By mid-November more than 65 percent of the opened and unopened cocoons had been lost. Therefore, in order to obtain the best sample of cocoons to determine incidence of mortality factors and other population parameters such as sex ratio and fecundity, it is important to sample about the last week in September. Collecting during this period will insure that parasites have had adequate time to exploit the cocoon population and that few sawflies, if any, have emerged.

Often some cocoons from previous generations will still be on the host trees and understory vegetation. They can almost always be separated from new cocoons, because cocoons that have weathered a year or more are a darker brown than cocoons of the current generation, which are a light tan.

The best estimates of population parameters such as percent sawfly emergence, adult sex ratio, mean fecundity, percent parasitized, and percent fungus-killed sawflies can be determined by sampling the entire crowns of trees within the stand. However, this requires that sample trees be felled. Our experience has shown that reasonably good estimates of population parameters can be determined by sampling the lower crowns of fringe trees along the beach without felling sample trees. Cocoons are handpicked from host branches and placed in perforated or screen-bottomed plastic vials. In order to evaluate cocoon population densities, the amount of time in hours and minutes actually spent removing cocoons from foliage should be recorded. This enables computation of cocoons collected per man hour, a rough index of cocoon population density. It is important to remember, however, that cocoon population levels are not necessarily closely related to larval population levels, because fungus disease often decimates high larval populations before many cocoons are spun.

Unopened cocoons should be placed individually in gelatin capsules[2] to keep each sawfly or parasite with its cocoon; and incubated at 12° to 19° C (55° to 66° F) and 80- to 90-percent relative humidity. Cocoons to be held for delayed emergence must be incubated at temperatures above freezing but below 5.5° C (42° F), because sawflies can complete development and emerge at the warmer temperature.

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[2] No. 00 (9 x 25 mm) gelatin capsules are manufactured by Eli Lilly Company, Indianapolis, Indiana. Mention of any product or company does not constitute endorsement or approval by the U.S. Department of Agriculture to the exclusion of others that may be suitable.

Sawfly and parasite adults begin to emerge from the cocoons after several weeks of incubation. Emerging insects can be easily differentiated because sawflies are "thick waisted" and have serrated or plumed antennae (fig. 12A, B), whereas parasites are "thread waisted" and usually have long threadlike antennae (fig. 13A, B, C). Normally, male sawflies begin to emerge several days earlier than the females. Sawfly sexes are separated on the basis of antennal shape--plumed in the male (fig. 12B) and serrated in the female (fig. 12A).

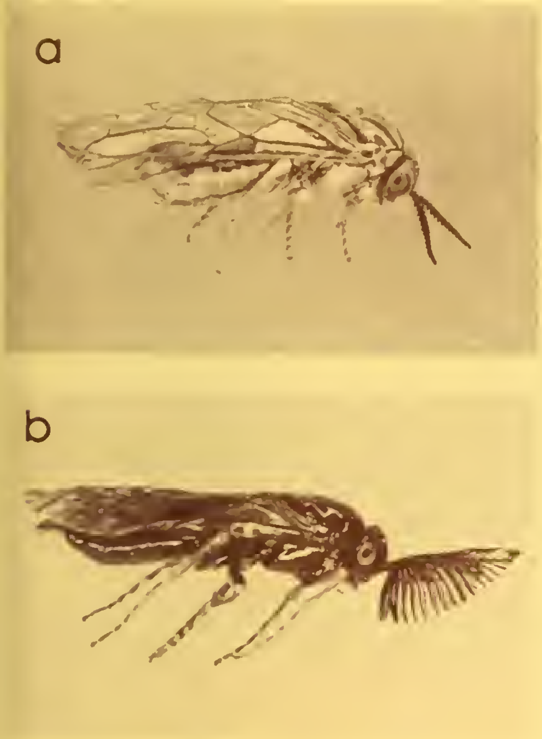


Figure 12.--Side views of adult hemlock sawflies showing: A, serrated antennae and light body color of female, and B, plumed antennae and dark body color of the male.

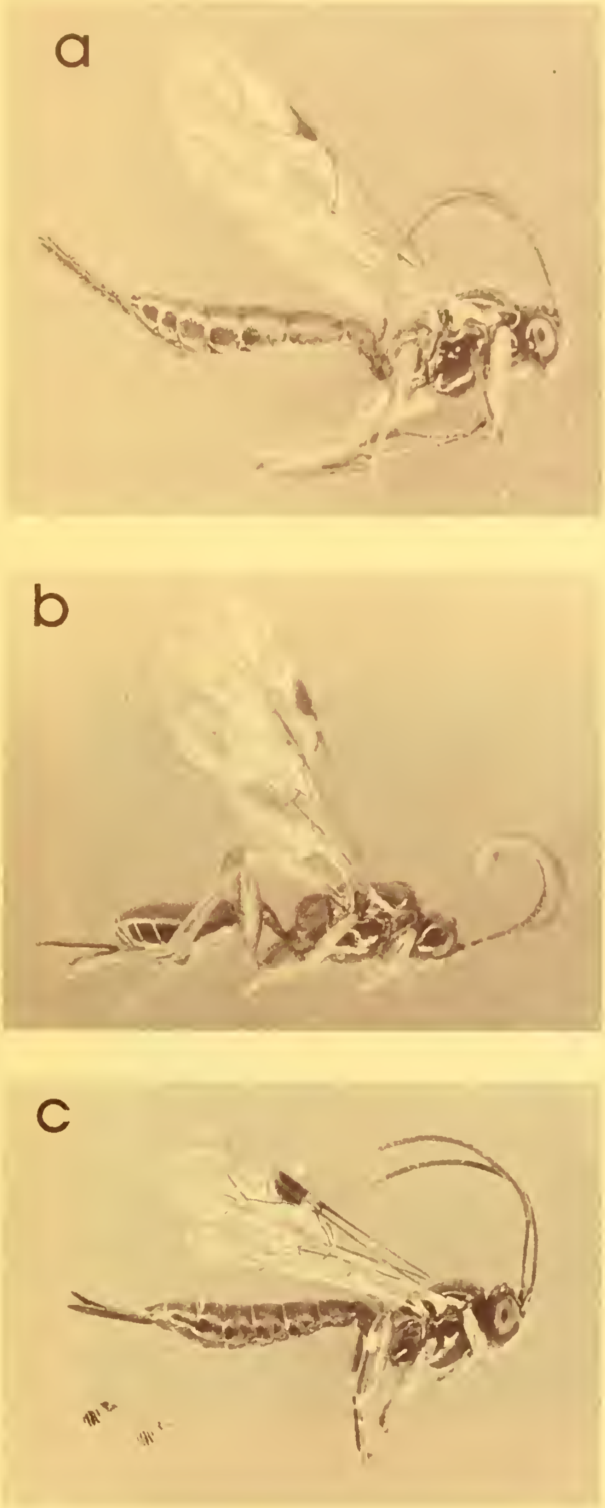
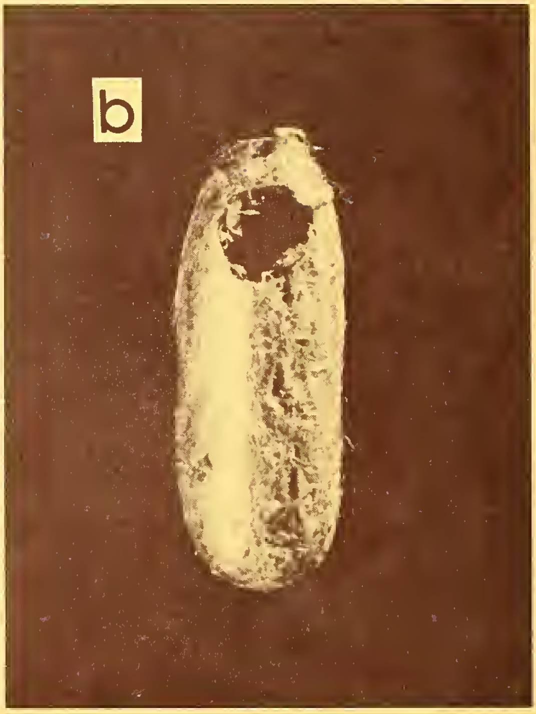


Figure 13.--Side views of adult females of: A, Delomerista japonica diprionis; B, Oresbius tsugae tsugae; C, Itoplectis quadricingulatus; *parasites that oviposit and emerge from cocoons of the hemlock sawfly.*

There are nearly 20 species of insects in about a dozen genera that parasitize the larva or cocooned prepupa of the sawfly. All the important sawfly parasites kill the host and overwinter within the host cocoon; parasite field emergence occurs in early summer the following year. Emerging parasites chew a jagged hole at or near the end of the cocoon (fig. 14A, B). Normally, only a single parasite emerges, but sometimes as many as a dozen tiny wasps emerge from one cocoon.

Overwintering for parasites is facultative so that parasitized cocoons collected in late September produce adults after several weeks of incubation at a temperature of 18° to 20° C (65° to 68° F).



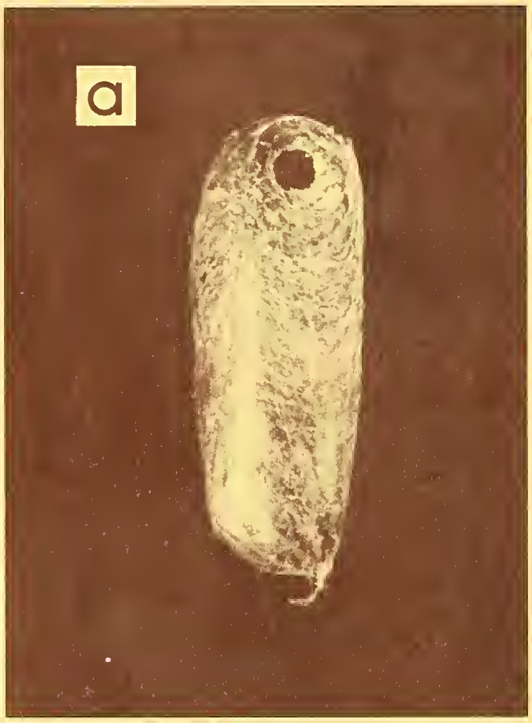


Figure 14.--Hemlock sawfly cocoons showing parasite emergence holes of: A, Amblymerus verditer; *and B,* Oresbius tsugae tsugae. *The latter is also characteristic of* Delomerista japonica diprionis *and* Itoplectis quadricingulatus.

The most commonly reared parasites are *Delomerista japonica diprionis* Cush., *Oresbius (= Opidnus) tsugae tsugae* (Cush.), and *Itoplectis quadricingulatus* (Prov.), which in combination with the rarer parasites in the complex, have accounted for up to 54-percent mortality in the cocoon population. The following key simplifies identification of the major adult parasite species.[3]

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[3] For a more complete treatment of sawfly parasite biology and identification, see Torgersen (1968).

Key to Some Parasites of *Neodiprion tsugae*

1. Wings absent, female.......................................*Gelis* sp.
1. Wings present, males and females.....................................2
    2. Forewing venation reduced (fig. 15); small (less than 3 mm), mostly metallic green-bodied wasps; short elbowed antennae; commonly gregarious..*Amblymerus verditer*
    2. Forewing venation well developed (figs. 16-22); body 5 mm or longer; antennae long and threadlike (fig. 13A, B, C).......................3
3. Forewings with dark bands (fig. 16); body and legs reddish brown with darker areas on thorax and abdomen........................*Gelis tenellus*
3. Forewings without dark bands (figs. 17-22)..............................4
    4. Areolet present (figs. 17-20)..5
    4. Areolet absent (figs. 21, 22)..8
5. Areolet five-sided; second intercubitus (fig. 17) sometimes faded; females with narrow light band along inner margin of compound eye; males with white face; female as in fig. 13B...................*Oresbius tsugae tsugae*
5. Areolet three- or four-sided (figs. 18-20)...............................6
    6. Areolet three-sided (fig. 18); first tarsal segment of hind leg about half as long as tibia; ovipositor very short, barely reaching beyond end of abdomen; face, clypeus, antennal base anteriorly and/or mandibles commonly patterned with yellow; lower portion of thorax sometimes yellow...*Lamachus* spp.
    6. Areolet four-sided (figs. 19, 20); first tarsal segment of hind leg less than half as long as tibia; ovipositor about as long as hind tibia...7
7. Inner margin of compound eye markedly concave opposite antennal base (fig. 23); hind tibia of both sexes black, with an encircling white band near basal one-third (figs. 13C, 25); wing venation as in fig. 19; face of both sexes black.............................*Itoplectis quadricingulatus*
7. Inner margin of compound eye not markedly concave opposite antennal base (fig. 24); hind tibia without encircling white band, usually white along ventral margin and/or basally (figs. 13A, 26); face of male white, face of female black; wing venation as in fig. 20....*Delomerista japonica diprionis*
    8. Radius and cubitus divergent (fig. 21); ovipositor short, barely reaching beyond end of abdomen.........................*Lamachus* spp.
    8. Radius and cubitus slightly convergent, then divergent (fig. 22); ovipositor long, about as long as hind tibia.............*Mastrus* spp.[4]

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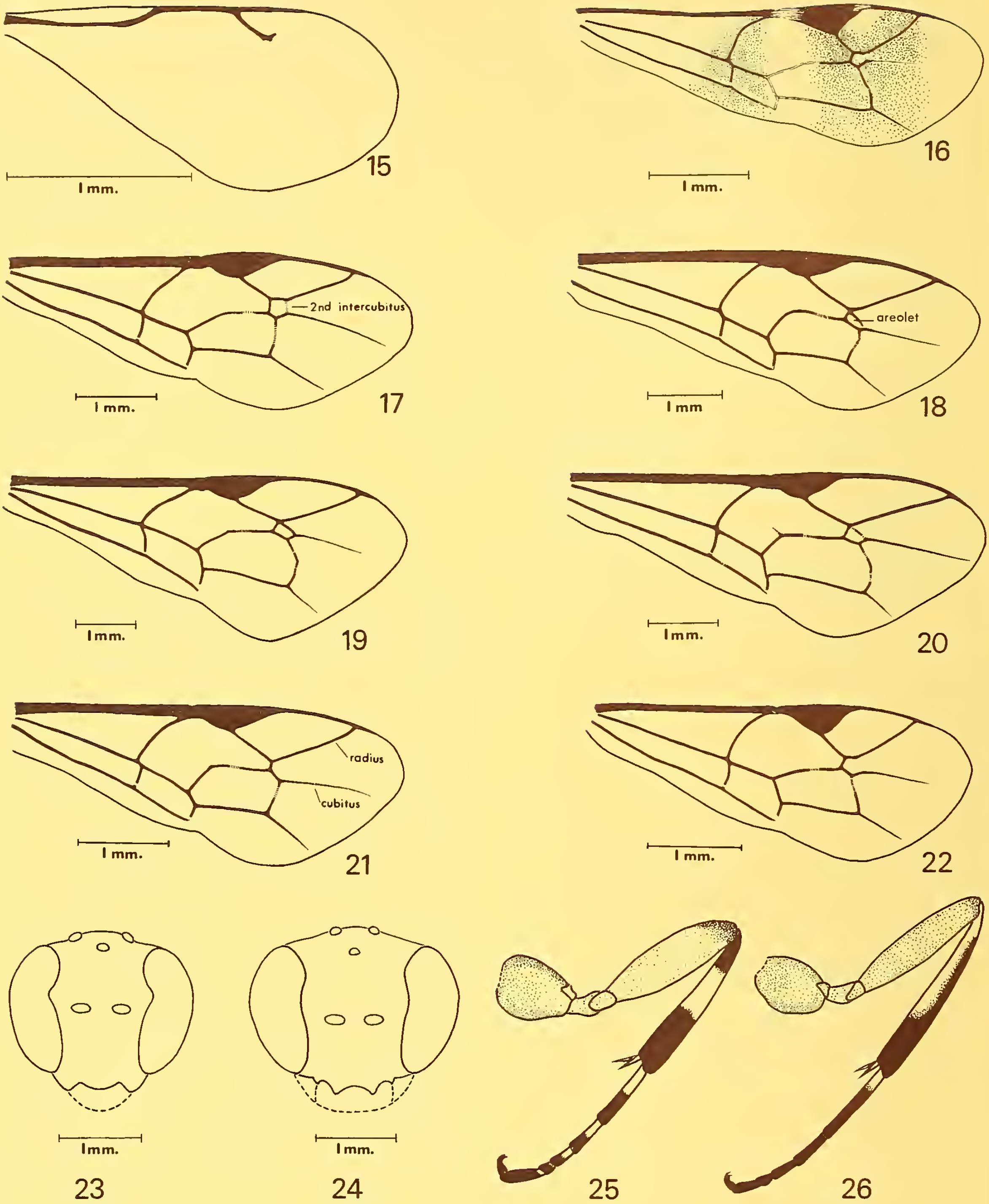
[4] Males of some uncommon *Gelis* species will also key out here.



Figures 15-26.--15-22, forewings: 15, Amblymerus verditer; *16,* Gelis tenellus; *17,* Oresbius tsugae tsugae; *18,* Lamachus *sp.; 19,* Itoplectis quadricingulatus; *20,* Delomerista japonica diprionis; *21,* Lamachus *sp.; 22,* Mastrus *sp. 23, 24, anterior view of heads: 23,* Itoplectis quadricingulatus; *24,* Delomerista japonica diprionis. *25, 26, hind legs: 25,* Itoplectis quadricingulatus; *26,* Delomerista japonica diprionis.

When sawfly and parasite emergence has ceased, remaining cocoons should be dissected and their contents analyzed. Contents of dissected cocoons will fall within categories such as the following: unemerged sawfly or parasite adult, sawfly or parasite pupa, prepupal sawfly larva in extended diapause, parasite larva, and *Entomophthora*-killed sawfly larva. Most categories are readily determined through ocular examination, but determination of the presence of *Entomophthora* or of a parasite larva requires special procedures. Usually, the *Entomophthora*-killed host consists of a dried, mummified, barely recognizable larva within the cocoon. A smear slide can be prepared by placing a bit of tissue in a drop of water or 10-percent potassium hydroxide (KOH) solution on a microscope slide and pressing a cover slip into place. Examination of the tissue through a compound microscope at 150X magnification verifies the presence of *Entomophthora* if the smear appears to be composed of spherical, thick-walled resting spores 25 to 30µm in diameter (fig. 27).

Living parasite larvae are legless, tapered posteriorly, and cream colored. Dead parasite larvae become shortened, dark brown, and shriveled. Verification of the species of a living or dead parasite larva is based mainly

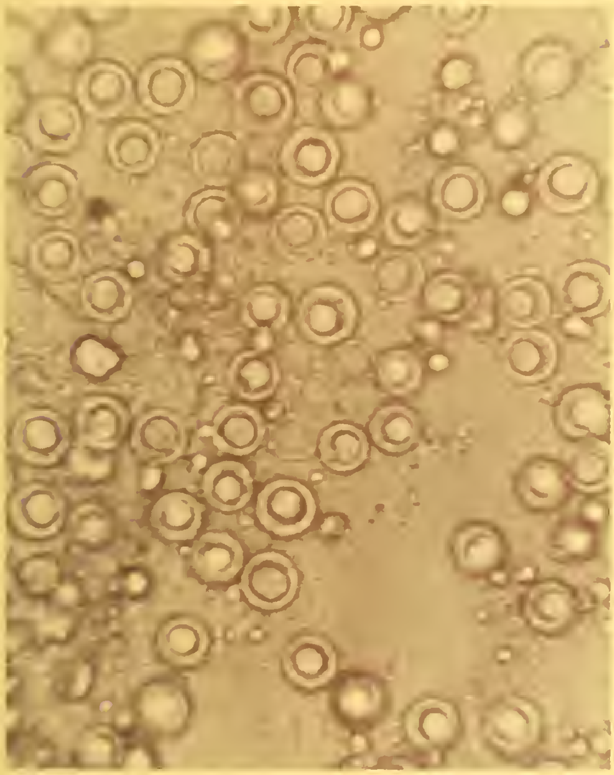


Figure 27.--Resting spores (270X) of Entomophthora sphaerosperma.

on the appearance of the sclerotic structures associated with the mouthparts. These structures (fig. 28A, B, C)[5] are apparent from the front on both living and dead larvae. Soaking the dead parasite larva in a 10-percent solution of KOH for an hour or more until the carcass is softened makes the larval skin easier to manipulate.

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[5] For a more complete treatment of identifications of the larval stage of sawfly parasites, see Torgersen (1969).

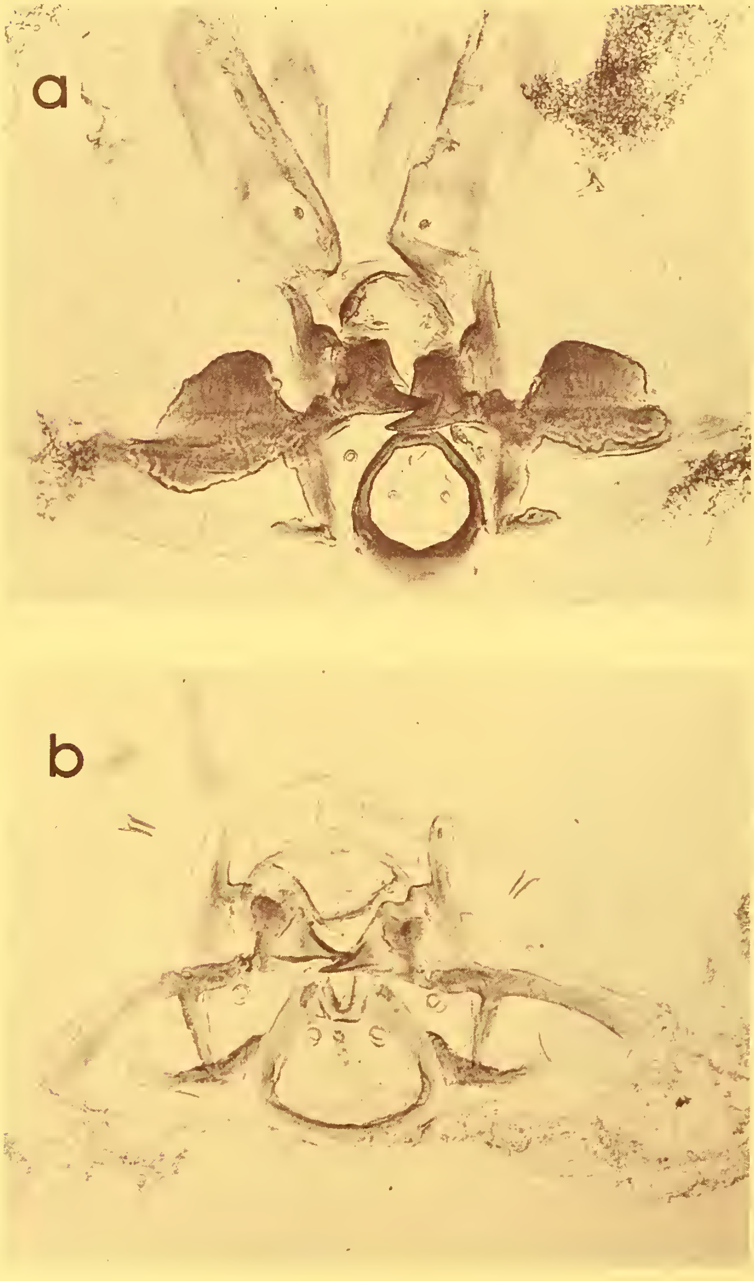

Figure 28.--Anterior views of structures associated with the larval mouthparts of: A, Delomerista japonica diprionis; *B,* Oresbius tsugae tsugae.

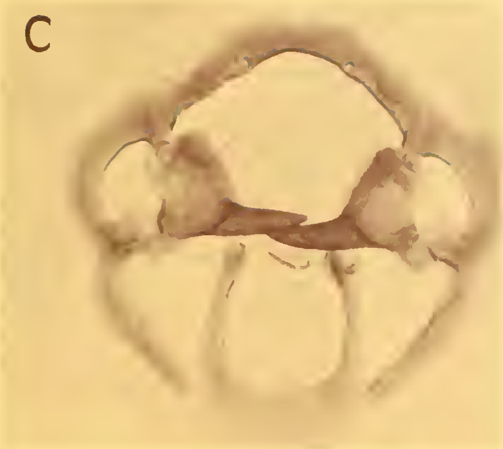

Figure 28.--Anterior views of structures associated with the larval mouthparts of (continued): C, Itoplectis quadricingulatus.

Determination of the parasite responsible for death of the cocooned sawfly is possible even if the host cocoon is separated from the parasite which emerged from it, or if opened cocoons are collected following field emergence of the parasites. Since fewer than 10 percent of cocoons remain on foliage or twigs beyond June of the season following their formation, most cocoons, whether opened or not, will be found on the ground. The presence of an emergence hole (fig. 14A, B) verifies parasitization, and dissection will reveal the nearly white, shriveled, shed larval skin with the characteristic mouthparts. An hour's soaking in KOH softens the dried skin to facilitate observing the mouthparts. Preparation of a semipermanent microscope slide using a nonresinous mounting medium[6] will also simplify examining the mouthparts for making a determination.

Diameter of all cocoons should be measured using a dissecting microscope fitted with a calibrated ocular micrometer disk. Measurements can be made when unopened cocoons are being dissected. The microscope, its magnification, and the precise conversion ratio of ocular micrometer units to millimeters used for measuring the cocoons should also be recorded. For accurate measurements, the microscope calibration should be rechecked before each use. Cocoon diameter measurements are useful because they provide a means to determine sex of cocoons that did not produce adult sawflies. This is done by plotting frequency distribution of cocoon diameters grouped according to arbitrary size classes of about 0.15 to 0.20 mm. Male cocoons are smaller than female cocoons, and overlap is minimal (fig. 29). Sometimes female cocoon diameters have a bimodal frequency distribution; but there is not as sharp a dip between the modes as there is between male and female cocoon diameters, which occurs between 2.6 and 3.0 mm. Cocoon size varies among sample areas and years; therefore samples from separate areas must be plotted separately to determine the separation point between male and female cocoons as accurately as possible.

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[6] Turtox CMC-10 can be used for this purpose. Available through General Biological Supply House, Inc., 8200 South Hoyne Ave., Chicago, Illinois 60620.

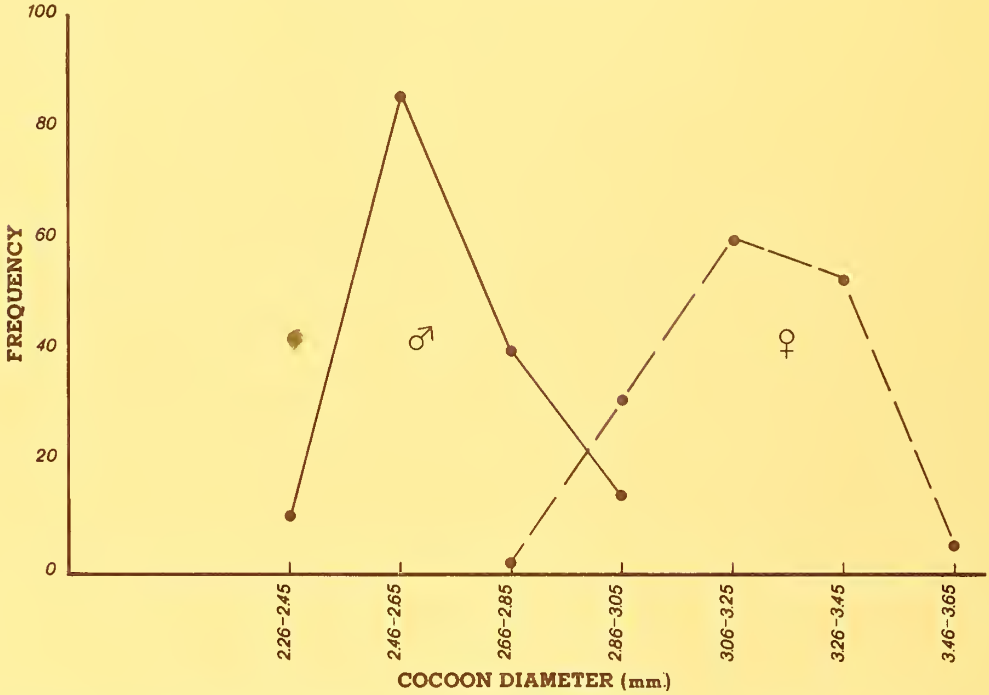

Figure 29.--Hemlock sawfly cocoon diameters (Hard and Schmiege 1968).

The separation of sexes can be hastened by sorting cocoons using slotted seed dockage sieves[7] (fig. 30). A tray with 2.98-mm x 19-mm (7.5/64- x 3/4-in) slots retains large female cocoons and passes smaller females and males. A lower tray with 2.58-mm (6.5/64-in) slots passes most male cocoons but retains intermediate diameter male and female cocoons. These must be measured and their frequency distribution plotted to determine the separation point between sexes. When the females emerge they can be either dissected immediately and eggs counted, or preserved in KAAD and dissected later to determine actual fecundity.

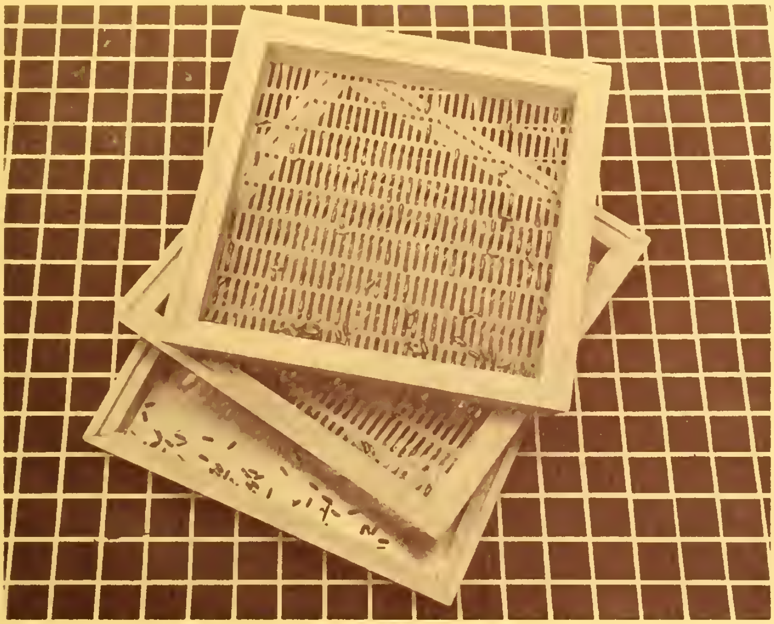

Figure 30.--Seed dockage sieves used for size-sorting hemlock sawfly cocoons.

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[7] Manufactured by Burrows Equipment Company, 1316 Sherman Avenue, Evanston, Illinois 60204--9- by 9-in perforated metal sheet type, oblong holes 6.5/64- and 7.5/64- x 3/4-in.

Female cocoon diameter can be used to estimate fecundity. However, the correlation and slope of fecundity on cocoon diameter varies among places and years, so the regression in figure 31 should not be used to estimate fecundities of new samples. A double sampling procedure requires measuring cocoon diameter and fecundity of a small random sample of females to determine the regression of fecundity on cocoon diameter. Then cocoon diameters of a large number of female cocoons are measured, and the mean diameter is entered in the regression equation[8] to estimate population

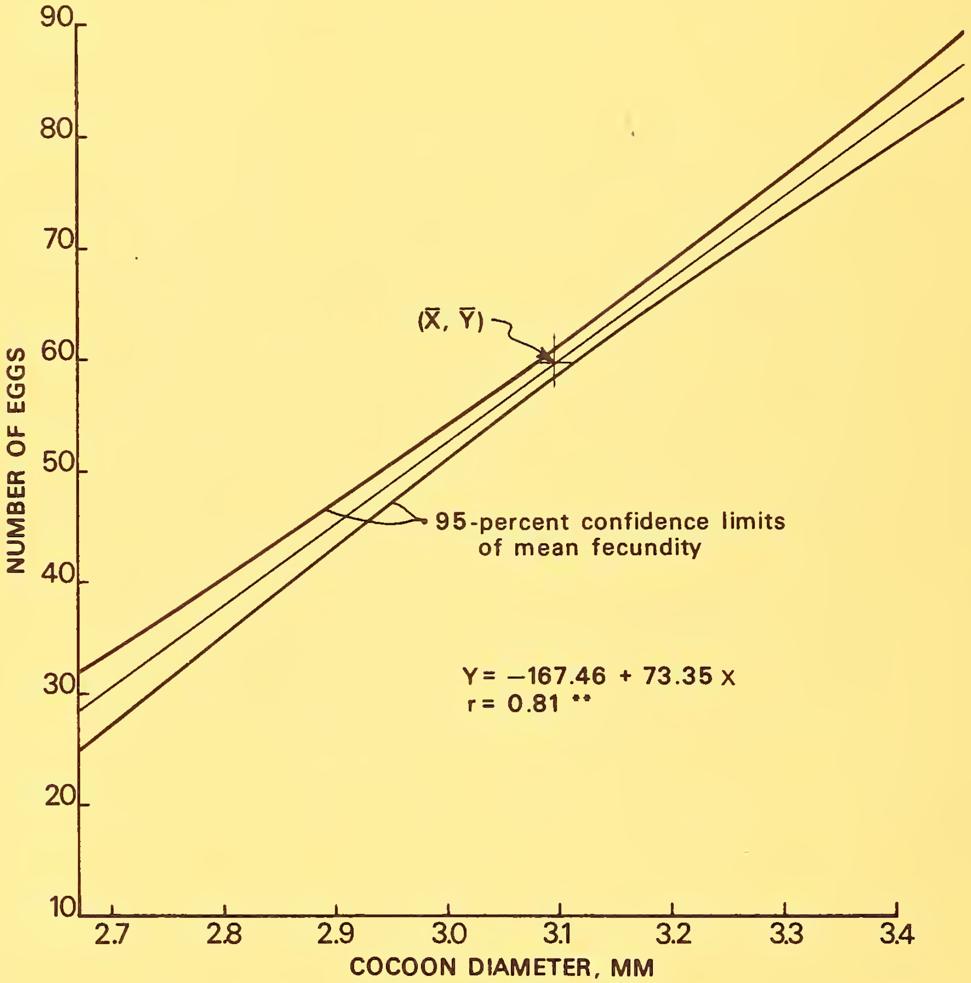

Figure 31.--Relation of hemlock sawfly fecundity to cocoon diameter, southeast Alaska, 1972.

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[8] The formula for estimating population mean fecundity, $\bar{Y}$, is $\bar{Y} = \bar{Y}_S + b(\bar{X}_L - \bar{X}_S)$, where $\bar{Y}_S$ = mean fecundity of the small sample, b = regression coefficient of the small sample, $\bar{X}_L$ = mean cocoon diameter of the large sample, and $\bar{X}_S$ = mean cocoon diameter of the small sample.

mean fecundity. Female forewing length is also highly correlated with fecundity (fig. 32); therefore double sampling may be used to estimate mean fecundity of a sample by establishing the regression of fecundity on forewing length of a small random sample and then use the regression to estimate mean fecundity of a large sample whose mean forewing length is known. The benefit of double sampling is that cocoons and wings of many females can be measured in the time required to dissect and count the eggs in a single female, and accuracy in estimating overall population mean fecundity will likely be improved.

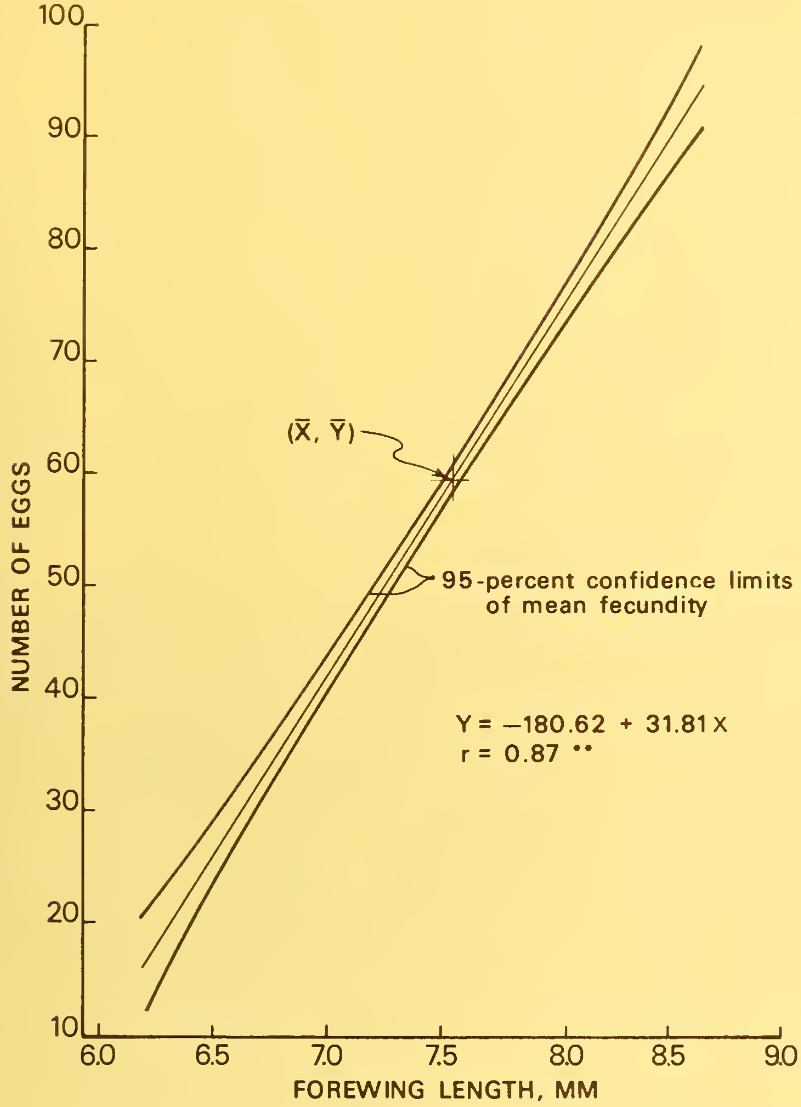

Figure 32.--Relation of hemlock sawfly fecundity to forewing length, southeast Alaska, 1972.

EGG SAMPLING

Field egg populations can be sampled after November 1, when most females have completed oviposition. During an unusually cold fall, females may have little opportunity to lay eggs. Days are short and often stormy in November, but fall egg sampling may be required to provide leadtime if control activities are anticipated for the following summer. Otherwise egg sampling can be postponed to the following March or April when days are longer.

If an infestation borders on a beach or clearing, an experienced entomologist can determine whether a high egg population is present by randomly selecting western hemlock branch tips reached from the ground and examining them for presence of egg clutches (fig. 33). The branch tips most likely to bear eggs are those that project farthest from the tree crown and are most exposed to the sun. This gives only a very rough index of egg abundance, however, and requires much empirical knowledge of the sawfly.

A better technique for assessing egg populations is through a sequential sampling plan[9] developed for this species. Intermediate crown class western hemlock trees are selected randomly. Four 18-in branch tips are removed from the midpoint of the upper crown of each sample tree and examined until an egg is found. A trace is drawn up one square on the sequential sampling form (fig. 34) for an egg-bearing tree or right one square for a tree without eggs. Trees are sampled sequentially until the trace drawn on the form crosses a decision line. The intermediate decision lines are adequate for most sampling because they can be expected to give an incorrect decision only 1 time in 10.

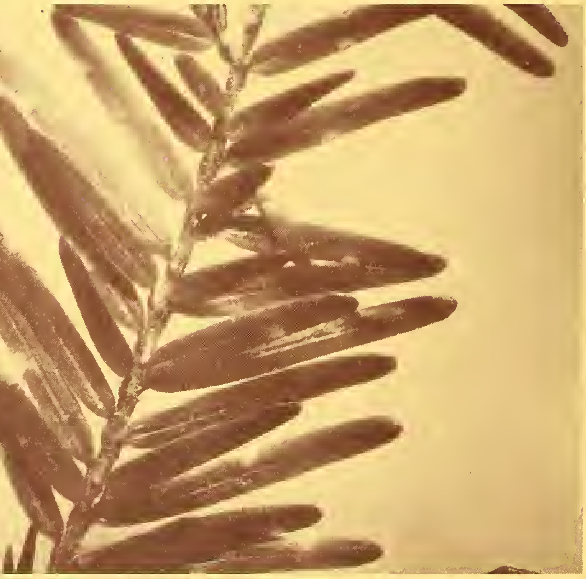

Figure 33.--Hemlock foliage showing sawfly eggs deposited in the edges of needles.

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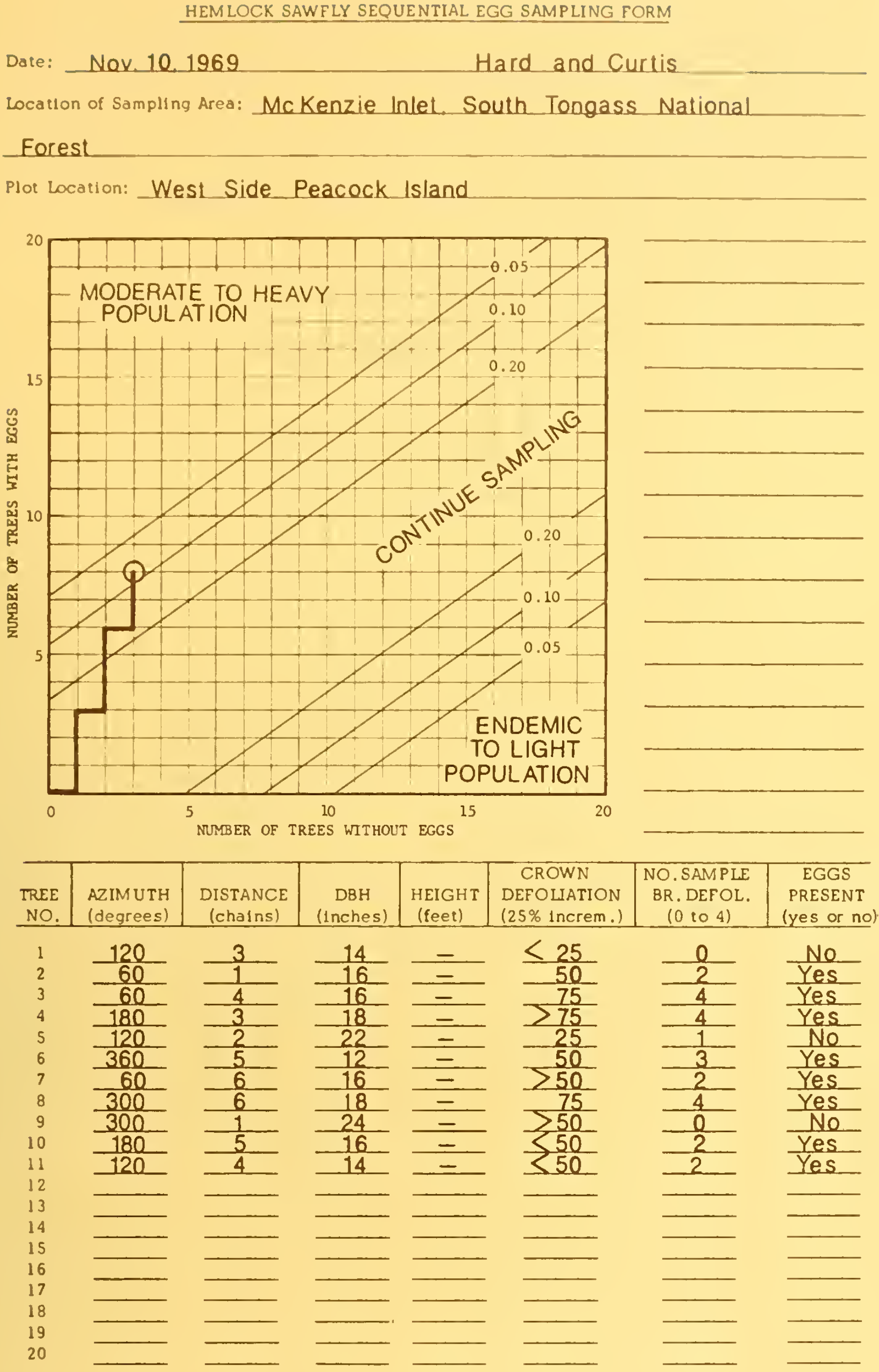
[9] For a more complete description of the sampling procedure, see Hard (1971b).



HEMLOCK SAWFLY SEQUENTIAL EGG SAMPLING FORM

Date: Nov. 10, 1969 Hard and Curtis

Location of Sampling Area: McKenzie Inlet, South Tongass National Forest

Plot Location: West Side Peacock Island

MODERATE TO HEAVY POPULATION
CONTINUE SAMPLING
ENDEMIC TO LIGHT POPULATION
0.05
0.10
0.20
0.20
0.10
0.05
NUMBER OF TREES WITH EGGS
NUMBER OF TREES WITHOUT EGGS
0
5
10
15
20

TREE NO.	AZIMUTH (degrees)	DISTANCE (chains)	DBH (inches)	HEIGHT (feet)	CROWN DEFOLIATION (25% increm.)	NO. SAMPLE BR. DEFOL. (0 to 4)	EGGS PRESENT (yes or no)
1	120	3	14	–	< 25	0	No
2	60	1	16	–	50	2	Yes
3	60	4	16	–	75	4	Yes
4	180	3	18	–	> 75	4	Yes
5	120	2	22	–	25	1	No
6	360	5	12	–	50	3	Yes
7	60	6	16	–	> 50	2	Yes
8	300	6	18	–	75	4	Yes
9	300	1	24	–	> 50	0	No
10	180	5	16	–	< 50	2	Yes
11	120	4	14	–	< 50	2	Yes
12							
13							
14							
15							
16							
17							
18							
19							
20							

Figure 34.--Hemlock sawfly egg sequential sampling form (Hard 1971b).

CONCLUDING REMARKS

Use of the techniques presented in this guide will enable pest management entomologists to predict the probable trend of hemlock sawfly infestations based on the following criteria:

1. Frequency of precipitation during the previous and current summers.
2. Population vigor of the current generation determined by adult sex ratio and fecundity.
3. Assessment of the effectiveness of mortality factors such as parasites and pathogens.
4. Expected larval population the following year determined by egg sampling.

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